Tested Studies for Laboratory Teaching
Proceedings of the Association for Biology Laboratory Education
Vol. 32, 78–87, 2011

The Role of Feeding Adaptations in Resource Competition between Invasive and Native Clams

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This laboratory allows students to investigate the feeding architecture of gills as an adaptation which may allow invasive clams to more successfully take advantage of local food resources, and thereby displace native clams. Students measure clearance rates in an invasive (such as *Corbicula* sp.) and a native clam (such as a unionid species) for two potential food items--algae and bacteria. The ability of these clam species to clear relatively large (algae) and/or small (bacteria) items is correlated with the measured spacing between cirri on gills. In addition to gaining experience in carefully controlling experimental design, participants refine skills in pipetor operation, microscopy, dissection and spectrophotometry.

Keywords: Invasive species, resource competition, gill, clam, bivalve

Introduction

This laboratory was created for a first year majors course in general biology. We restructured our introductory course for majors (Concepts in Biology; **http://www.emu.edu/biology-old/Concepts/Biol173.htm**) to focus on several specific themes to introduce students to fundamental concepts and issues in biology. For example, exploring the issue of invasive species is used for an introduction to ecology. This laboratory complements that section by giving students hands-on experience working with an actual invasive species, and by allowing them to investigate one factor important to the success of invasive species--competition for food resources. Two major goals of our introductory majors course are to build skills in good experimental design and in proper use of laboratory instrumentation. This exercise builds these skills through guiding students in experimental design and data interpretation, and by giving students experience with some key pieces of laboratory equipment (spectrophotometers, microscopes, and mechanical pipetors).

This laboratory was inspired by the studies of Silverman et al. (1996a,b), who have collected data showing how the feeding physiology of zebra mussels (*Driessena polymorpha*) and Asiatic clams (*Corbicula fluminea*) is different than that of native bivalves. Zebra mussels and Asiatic clams can capture particles less than 1 µm in diameter, suggesting that bacteria are a more important food source for these invasive clams compared with native unionid species. In general, Asiatic clams appear to be relatively non-selective in terms of the particles captured, successfully filtering algae of a variety of sizes (Boltovskoy et al., 1995). This stands in contrast to at least some native bivalves, which appear to be restricted to larger particle sizes (Riijsgard, 1988). In addition, invasive species having high rates of clearance (Sylvester et al., 2005) can have large ecological impacts via changes in algal concentrations (Phelps et al., 1994; Strayer et al., 1999). When algal concentrations are reduced, native clams may shut their valves and stop filtering (Riisgard et al., 2003). Thus, invasive clams may have a competitive advantage by starving out native clams while at the same time taking advantage of a resource (bacteria) that native clams cannot utilize.

This exercise tests the hypothesis that an invasive clam is able to take advantage of a food resource (the smaller-sized bacteria) that native clams cannot access. In the waters of our region, Asiatic clams have become very common and provide a good (and for this area, relevant) source of an invasive clam. We contrast the feeding physiology of these clams with native clams that are ordered from a commercial biological supply company.

The exercise is designed for two lab periods of 2 hours and 40 minutes each. In the first week of the lab, we spend considerable time initially talking about various reasons that invasive clams might be successful. We rely on students to come up with ideas based on their readings and on their general knowledge of biology. Through this guided discussion, we eventually talk about the potential role of food resources and feeding adaptations. After setting the background for the experiment, and clearly defining the hypothesis, we spend the remainder of the first lab period measuring algal clearance rates. Although the hypothesis is the same for all groups, students must work in their groups to design the methodology (for instance, deciding on one or more good controls for the experiment). In the second lab period, students continue with the methodology developed in the first period to assess filtration rates of bacteria. Finally, students dissect clams and use microscopy to assess cirri spacing in gills in order to establish a possible anatomical correlate to physiological differences which were observed.

Student Outline

Goals

- To investigate one aspect of invasive clam feeding ecology contributing to their success as invaders of waterways, displacing native clams.
- To practice experimental design, with an emphasis on appropriate controls and data interpretation.
- To help develop accurate pipeting skills.
- To learn principles and practice of spectroscopy, as applied to measurements of bacteria and algal concentrations.

Before lab read: McMahon, R.F. (2002). Evolutionary and physiological adaptations of aquatic invasive animals: r selection versus resistance. *Can. J. Fish. Aquat. Sci*. 59: 1235-1244. Focus especially on the sections "Life History Traits of…" and "Synthesis".

Before starting this lab, you should list common adaptations which characterize invasive species. Why would these characteristics allow an invasive bivalve the ability to outcompete native bivalves?

Introduction

Invasive species are a significant, but often under-appreciated, cause of alterations of both terrestrial and aquatic ecosystems. Watersheds throughout North America have seen alterations in their physical characteristics, and in the assemblage of species that are present, due to the accidental or intentional introduction of non-native species. New species continue to be introduced to waterways, further threatening efforts to conserve native species and to maintain ecosystems. Determining factors that contribute to introduced species becoming successful invasives is key to preventing the detrimental effects associated with the introduction of non-native species.

In order to thrive in a new habitat, an introduced species must either fill a previously unoccupied niche, or (more commonly) it must outcompete species that occupy an existing niche. Since communities usually have evolved such that species fill available niches, an introduced species generally becomes invasive when it successfully displaces a native species already present. In order to displace an existing species, an invader must possess characteristics that provide it with a competitive advantage over native residents. Ecologists have identified a number of possible characteristics that allow some species, such as the Asiatic clam and Zebra mussel, to outcompete native species (McMahon, 2002). Some of these traits are behavioral, while others are physiological. For instance, the ability to filter large volumes of water and to effectively remove food particles appears to be important to the success of these two invasive bivalves.

The Asiatic clam (*Corbicula fluminea*) is a good example of a non-native bivalve which has recently become nuisance species in many waterways of North America. Originally introduced to the west coast of North America from Southeast Asia in 1938, this species has spread across the continent and is now found in most major waterways, preferring running water with a sand or gravel substrate. In addition to having significant economic impacts, these bivalves have outcompeted and displaced native bivalve species. Asiatic clams can have significant ecological effects, such as causing large decreases in algae concentrations of rivers (Phelps, 1994; Cohen et al., 1984). Although the Appalachian region was once known for its diversity of bivalves, native species are now generally rare.

Figure 1. Adult Asiatic clam (*Corbicula fluminea*).

Asiatic clams have an anatomy that is typical of bivalve molluscs (literally "two shells"). When filtering, the clam opens its shells several millimeters and circulates water through the shell cavity via the incurrent and excurrent siphons. Food and oxygen are thus brought into contact with the gills, while waste products are removed. Water flow occurs by the action of cilia on lateral cells of gills. Capture of food occurs by a different set of cilia, the eulateral frontal cirri (EFC). These cilia are fused together and located on laterofrontal cells which are located between the lateral cells and the front of the gill (Silverman et al., 1996). Water passing between gill lamellae therefore are trapped by these cirri, and subsequently transported by the gill mucus to the mouth of

the clam. These cirri either physically trap the particles themselves or create complex water currents that trap the material (see Silverman article).

In this lab you will attempt to measure aspects of feeding in both an invasive bivalve (such as *C. fluminea*), and in a native clam (such as *Anodonta* sp.). You will observe the general feeding behavior of the clams, measure the feeding rates of these clams on both algae and bacteria, and then make some morphological measurements of the feeding apparatus of these animals. In the process, you will learn to use several pieces of instrumentation that are critical to your success in the biology laboratory.

Data for this investigation will be collected over a two-week period. In the first lab period, you will learn how to measure filtration rates in clams, and apply that to estimating algal filtration rates. In order to do this you will learn how to use a spectrophotometer, an instrument that is key to laboratory experimentation in biology. In the second lab period, you will measure filtration rates of bacteria and determine the cirri spacing in the gills. These measurements should allow you to compare the utilization of these two food sources with the anatomy of the food-gathering structure of clams (the gills).

First Lab Period: Measuring Algae Clearance

We will measure how fast clams remove algae from water, using cultures of algae that are representative of the normal food items of bivalves. For instance, two freshwater species of algae that may be used are *Ankistrodesmus* sp., and *Chlorella* sp. *Ankistrodesmus* sp. is an elongated alga with dimensions of approximately 2 x 30 micrometers (µm). *Chlorella* sp. is a spherical alga with a diameter of approximately 5 µm.

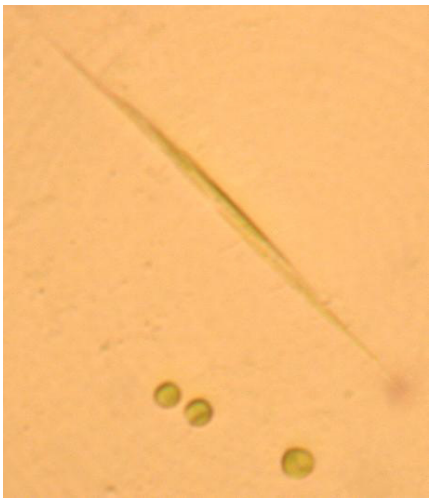

Figure 2. Light micrograph of *Ankistrodesmus* (elongated) and *Chlorella* (spherical).

Make sure you read through the instructions before starting the experiment. The instructions describe decisions that need to be made regarding your experimental design and contain the details on the measurement techniques; refer to this section as you plan your experiment.

Measuring Algae Concentrations with Fluorescence Spectroscopy

The standard method of assessing algae concentrations in natural waters is to measure chlorophyll. Chlorophyll concentrations in water are generally proportional to algal biomass (total mass of algae in a sample). Chlorophyll, the light-harvesting molecule in plants that accounts for their green color, is measured using a technique called fluorescence spectroscopy.

Experimental Design

The native clams are a different size than the invasive clam. This represents a confounding variable that we wish to account for. How do we do adjust for the difference in size? Often, we will "standardize" the measurement to a unit of mass or length. For our purposes, it's easiest to keep the volume proportional to the relative weight of the clams. For example, if the native clam has a mass 4x greater than the Asiatic clam than the volume of solution should be 4x greater in the native clam beaker.

As with any experiment, we need to have an appropriate control, and there are several possible controls that can be designed for this experiment. You should think of one control that you think is necessary, and then check with the instructor about whether it is appropriate.

Method

The technique for measuring algal clearance is as follows:

1. Make a 50:50 mix of dechlorinated water : algal culture. Before pouring from algal culture, let the big chunks settle to the bottom first so that they are not in the sample. Be careful when pouring not to stir up these big chunks. The total volume should be about 25 ml per gram of clam weight; calculate the volumes you need based on this and on the discussion above about standardizing conditions.
2. Get one of the clams to be used and carefully place it in your experimental beaker. These clams were held for 24 hours in water without food so that they could clear their guts of any material. (Production of feces or pseudofeces during the experiment can throw off the measurements).
3. Take one sample from each beaker as you are starting the experiment. When exactly do you think is the best time to take this sample? For each sample that you take for measuring chlorophyll concentration, you should use the P5000 pipetor to remove 2.5 ml of solution. Place the sample in a cuvette and measure fluorescence (or hold in the dark until all samples are collected and ready to measure), and then gently pour the sample back into the beaker.

Figure 3. Experimental setup showing *C. fluminea*

4. Watch for the clam to open its shell and begin siphoning. The incurrent and excurrent siphons are located on the posterior side of the clam. Once the clam opens, start the time course of your experiment. If the clam does not open within 10 minutes, it probably will not—get another clam.
5. Take at least three additional samples during the course of the experiment (for instance at 5, 10 and 20 minutes). The longer you let the experiment go, the better results you will likely get.
6. At the end of the experiment, weigh your clam.

Data Calculations

1. You now have a measure of chlorophyll content in the water at each time point. How would you process and plot these data? First plot the data, simply using the fluorescence values at each time point.
2. Normally in feeding experiments we would like to calculate a *feeding rate*: a value that expresses feeding as mass of food per time unit per mass of animal. There are four values you need for calculations: time, volume of fluid used, mass of clam, and fluorescence. Fluorescence is expressed as a unit-less value, but can be thought of as the *concentration* of algae present. From the concentration of algae and volume of fluid, you can calculate the mass of algae removed.

Second Lab Period: Bacteria Clearance and Cirri Spacing

The measurement of bacterial clearance is similar to that for chlorophyll. A volume of bacteria will be added to a beaker with a clam, and the number of bacteria followed over the course of the experiment. Instead of fluorometry, you will use absorbance spectrophotometry. As with the measurement of algae, you need to think about the use of experimental controls. What control did you use for the algae experiment? Do you need a similar control, or a different one for the bacteria experiment?

Measuring Bacterial Density with Absorbance Spectrophotometry

There are several ways of measuring bacterial density, including plating of samples on petri dishes and counting colonies. A faster way to measure bacteria is to measure the "optical density" (O.D.) of a solution. Optical density is simply how cloudy a solution is; for bacteria we shine light of a certain wavelength (550 nm) through the sample and measure how much light makes it through. The less light can pass through, the more bacteria (the cloudier) in the solution. Use of the spectrophotometer will be a very common operation in your biology courses, so you should become comfortable with its use at this point.

Procedure for Measuring Bacterial Density

The procedure for measuring bacterial clearance is very similar to that for algae; you can essentially follow those same instructions. Instead of adding algae, obviously you want to add bacteria for this experiment. **Take care in working with bacteria to avoid contamination by keeping bacteria contained in your work area and wearing gloves**.

1. We have a sterile culture of *E. coli* growing to use as a source for those bacteria. Fill a 50 ml centrifuge tube with the culture and spin it at 3000 rpm for 5 minutes. Make sure the centrifuge is balanced when you do this.
2. **Gently** remove the centrifuge tube from the machine, there should be a loose "pellet" of bacteria at the bottom of the tube when you have finished. Gently pour off the brownish broth (the fluid left after centrifugation of any sample is referred to as supernatant), leaving the pellet at the bottom.
3. Add about 1 ml of dechlorinated water to the tube and shake it to resuspend the bacteria. Now pour this into a larger beaker containing enough dechlorinated water for your experiment.
4. Take a 2 ml sample from the beaker into a cuvette and measure its absorbance. You should get absorbance between 0.100 and 0.200. Spin more bacteria to add if your absorbance is low; add more dechlorinated water if the sample is high.
5. From this bacterial solution, take the required amounts of fluid for your experiments on the clams. Take a sample every 10 minutes for measurement. Measure this sample immediately and then pour it back into the beaker with the clam (keeping the volume equal throughout the experiment).
6. Calculate and plot bacterial filtration rate as you did for the algae samples last week.

Measuring Cirri Spacing

Clams capture food in part because of the presence of structures called cirri on their gills. Gills are made of many filaments, each of which contains rows of cirri. The cirri, which are bundles of cilia (cilia are the small hair-like structures common on microorganisms, and many animal tissues), wave back and forth. This action both pumps water between the filaments of the gill, and also allows for capture of particles. We wish to see if the spacing between cirri on the gill of these clams correlates with their feeding habits. Would you predict any difference in cirri spacing?

Dissect one of the *Corbicula* in order to obtain a piece of gill. Use the dissecting microscopes to remove the gill. Each side of the clam has two "demibranchs" (gill layers). Place a piece of gill on a slide with a drop of water and a coverslip. Examine this under the compound microscope with the 40x objective. A single native clam will be dissected for the class, with gill pieces distributed to all groups. Make a slide of this gill for analysis.

Once you are convinced that you have a good sample, have your instructor take you to a microscope with a digital camera to take a picture of the gill. Use this photograph to measure the distance between cirri by comparing it to the picture of the ruler markings.

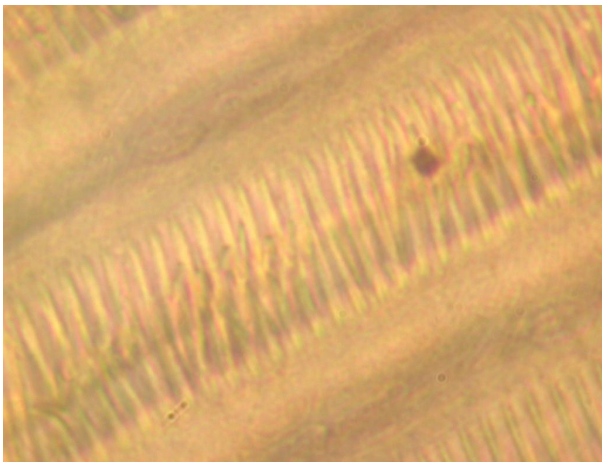

Figure 4. Image of eulateral frontal cirri from *Corbicula* gill, taken with 40x objective.

Materials

Day 1: Algae Filtration

Equipment

- Fluorescence Spectrophotometer *or* microscopes and slides for algal counting
- Micropipettors (P5000) with matching tips
- Balances for weighing clams

Supplies (each group)

- 2-3 clams each of invasive and native species
- 1 beaker for each clam, size depending on clam size
- graduated cylinder(s), matching beaker size(s)
- Dechlorinated tap water, or spring water
- Cuvettes for spectrophotometer
- Cuvette holder

Day 2: Bacterial Filtration

Equipment

- Absorbance Spectrophotometer
- Micropipettors with matching tips (P5000)
- Centrifuge
- Balances for weighing clams
- Microscope with digital photography capability

Supplies (per group of two or three students)

- 2-3 clams each of invasive and native species
- 1 beaker for each clam, size depending on clam size
- graduated cylinder(s), matching beaker size(s)
- Dechlorinated tap water, or spring water
- Bacterial cultures
- Two 50 ml centrifuge tubes
- Bleach bottles
- Trays for containing bacterial work
- Gloves
- Cuvette holder
- Cuvettes for spectrophotometer

Notes for instructor

Before Lab (starting 1 week in advance)

The most important consideration in planning ahead for this lab is to ensure that sufficient algae are grown. This process is normally started several weeks prior to the lab, under controlled light and temperature conditions and in Algal-Gro (Carolina) media (sterile), usually on a shaker bath. We have grown *Ankistrodesmus* and *Chlorella*, which allows students to visually observe (under the microscope) algae with two very different shapes. Many algal species can be used, but we've had the most luck with these two species. *Chlorella* tends to grow to a higher density. Older cultures (e.g., 2 weeks) tend to end up with clumps of algae on the bottom which need to be thoroughly dispersed before use. In the event that our cultures crash or we run out of algal culture, we sometimes take water from the pond in our greenhouse, which typically has standing growth of algae. Although not optimal, it does make a nice "emergency backup" for situations when we need more algae. We've found that using water from outdoor ponds can be difficult, as there is often suspended sediment and detritus that makes it difficult to work with. We rarely have cultures that have too much algae; more commonly the fluorescence measurements are somewhat low which gives a lower signal to noise ratio. Bacteria (*E. coli*) are normally grown 1-2 days in advance, in sterilized test tubes containing bacterial media.

Clams can be obtained from a number of sources. In many areas of the U.S. and Canada, *Dreissena* or *Corbicula* are collected from local waterways, where they occur in large abundance. We collect *Corbicula*, which are common in areas of sandy bottom and just upstream from ripples. Collection is easiest using a shovel and mesh screen; river substrata is pushed with the shovel in the direction of water flow, onto the tilted screen which then collects the larger items. Larger clams work best for the lab. Because native unionid clams are extremely rare in many regions, they are often easier to order (e.g., from Carolina Biological Supply) 1-2 weeks in advance of the lab. Clams should be starved for 2-3 days prior to the experiment. This has two functions: hungry clams are more likely to open and filter during the experiment, and starved clams do not produce copious pseudofeces which interfere with measurements.

Day 1: Prelab Preparation for Students

For the best results as a learning exercise, it is important that students read the background material, and think about possible reasons for the success of invasive species. We emphasize this to students prior to the lab, and have reasonable success in engaging the students in an informed discussion at the start of the lab. We lead a discussion where we specifically elicit ideas from students about what the characteristics are of a successful invasive species. Often we have them brainstorm in small groups before talking about this as a large group.

Following are items which come out of the discussion as possible ways that invasive clams might outcompete native clams. Much of this comes from the McMahon article which they are to have read before the class. We usually mention them if students have not identified them in the discussion.

- polyphagous (wide feeding niche)
- short generation times
- genetic variability
- rapid growth rates
- high dispersal rates
- single parent reproduction (vegetative)
- phenotypic plasticity
- large native range
- able to tolerate and function in a wide range of physical conditions

Further discussion time is used to clarify the difference between K-selected and r-selected species, and the life cycle of clams. Specific adaptations of *Corbicula* (as an example of an invasive clam) that are postulated to give them a competitive advantage are:

- higher filtration rates, and higher assimilation rates
- most assimilation is devoted to reproduction and growth
- juveniles can resuspend to be dispersed
- hermaphroditic and self-fertilizing
- tolerant of suspended silt
- high metabolic rates allow burrowing rapidly
- byssal thread allows transport by birds
- pediveligers can crawl upstream

The sea grant program has a bulletin which describes *Corbicula* and which serves as an example of the types of characteristics that allow for successful dispersal (**http://seagrant.wisc.edu/Home/Topics/InvasiveSpecies/Details.aspx?PostID=659**).

This next leads into a discussion of what bivalves eat, and the difference between algae and bacteria as food sources. From this we prompt students to think about how a hypothesis about feeding differences could be tested, which leads into a discussion of gill structure and experimental design.

Day 1: Algae Clearance

Students sometimes have problems with clams opening up and filtering. We ask them to watch carefully for activity on the shell edge and at the siphons, and start the filtration period once the see filtration occurring. We find that variability in valve opening is associated with individual clams, and therefore have them change clams if they do not see the clam opening within 10-15 minutes. We therefore keep as many clams on stock as possible, so that students have plenty to choose from. Once filtration starts, it's important that the experiment runs at least 20-40 minutes. Algal and bacterial measurements can be variable, so that getting a clear signal sometimes takes at least this amount of time. As mentioned earlier, experiments work best when the gut is cleared by fasting for several days prior to the lab. Gut clearance by clams during feeding will increase turbidity in the water, causing spurious results. The production of these pseudofeces does provide an opportunity to talk about the potential role of bivalves in nutrient cycling, since pseudofeces production is likely an important component of nutrient cycling in some ecosystems (such as oyster beds; see Silverman et al., 1996 for a discussion of this). Likewise, students need to be aware that material on the shell (e.g., algae) which gets in their sample will affect absorbance readings. We typically prompt them to think through this and solve the problem on their own, rather than tell them directly how to deal with these issues in experimental design.

A key feature of how we run this lab is to generally *not* to tell students everything about the lab. In particular, there are two aspects to this part of the lab that we try to get students to work out for themselves, and that they often find difficult. First, working with experimental design can be a challenge. We try to have students think through aspects of the design, such as the use of controls, and when/how samples are taken. Although difficult, we find this is an important part of the experience for students. For instance, it's important that students understand the role of the controls. A good example of this to have students understand that the "disappearance" of algae or bacteria from the water column might not be from filtering, but rather could be because of settling. If students do not think of it themselves, we make sure that by the end of the lab they understand that a good control would be to measure algae or bacteria in a beaker *without* a clam present.

The second challenging aspect can be how to work with the data. We find it important that students think about what the fluorescence or absorbance results actually mean. We often step through the example of what results (in algae concentration) we would get if equal volumes of water were used for two clams of different sizes that are filtering at equal rates relative to their mass. In that case, the faster rate of algae disappearance from the beaker with the larger clam makes it appear that it is filtering more effectively. However, on a mass-specific basis (per g tissue), they would be filtering at equal rates. In the procedure, we place clams in water volumes that are proportional to their mass, which simplifies the data processing for students (since rate of disappearance then would be indicative of filtration efficiency). We do have students calculate a mass-specific feeding rate, which requires them to think through some of these issues.

For a more advanced class, this can provide a good opportunity to discuss issues of scaling. For instance, from principles of allometric scaling we would not actually expect the rate of filtration directly scale with mass. Students with background in scaling could apply these principles to the data calculations.

We've found that fluorescence measurements of chlorophyll can be sensitive to specific conditions, and at times have given highly variable results. If this happens, or if a fluorometer is not available, an alternate method to estimate algae concentration is simply to count algae under a microscope. Likewise, a particle counter presumably could also be used, although we have not tried this.

Day 2: Bacteria Clearance and Cirri Spacing

To avoid effects of adding nutrient broth to the beakers with clams, we have students spin bacteria for about 5 minutes at ~3,000x. The exact conditions for spinning can vary with bacterial culture, ideally we want to spin the bacteria down without getting them too clumped together (when they are difficult to resuspend). Two 50-ml centrifuge tubes that are spun normally gives enough bacteria to give measurable absorbance (550 nm) when added to 500 ml of water. Media is poured off, and the bacteria are resuspended in dechlorinated tap or spring water for use with the clams. The use of *E. coli* in the experiments entails that proper safety precautions are followed. Gloves are provided, and all bacteria handling is done in a tray to limit the spread of the bacteria. Bleach bottles are provided to clean surfaces. Bacteria are autoclaved following the experiment.

We try to have every group do a clam dissection so that they get exposure to clam anatomy related to the filtration that they observe. This works well with *Corbicula*, since we typically have plenty of this species. We typically do not have many native clams (since they need to be ordered), and dissect one for the entire class. A single gill has plenty of tissue to distribute among multiple groups. We encourage students to look at the overall gill structure under the microscope; the gill is a fascinating organ and usually has intrinsic interest for students. For instance, cirri action is typically vigorous, and in fact can cause the gill to "walk" across the slide. Also, this lab is typically run in early fall, when pediveligers are present in *Corbicula* gills (Fig. 5), which stimulates discussion of the clam life cycle and generally helps engage students. Make sure that students are looking at frontal cirri. In giving directions for dissections, we usual refer students to one of the many websites that give instructions on clam dissection (for example, **www.biologyjunction.com/clam_dissection.htm**).

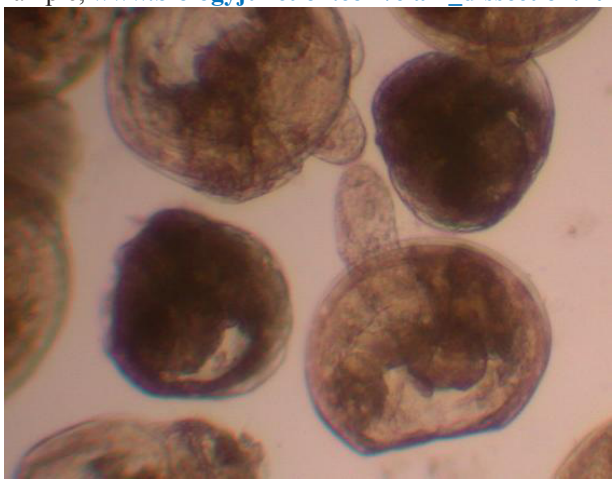

Figure 5. Immature Asiatic clams in the pediveliger stage; several are shown with the foot extended.

Conclusions and Sample Results

Students typically find that there is a correlation between the ability of clams to filter bacteria (the smaller food item), and the spacing in the cirri. Algal filtration rates tend to be similar in both clams, consistent with the larger size of these food items. Students thus find that the ability of *Corbicula* to take advantage of this food resource is a potential explanation for their success in waterways. The relevance of this is emphasized by the observation that native unionid clams are endangered in the Shenandoah Valley (and, in fact, very difficult to find!) while *Corbicula* are extremely abundant. This correlates with the lowered water quality in the region, often resulting in high bacterial populations (thus providing a food source for *Corbicula*).

Results for Algal and Bacterial Clearance

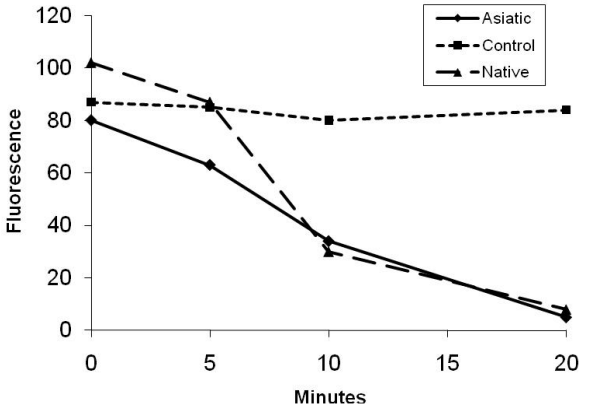

Figure 6. Algal clearance in native and invasive Asiatic (*Corbicula*) clams. Algal density is indicated in arbitrary fluorescence units (from chlorophyll fluoresence). Control condition is beaker without clams present.

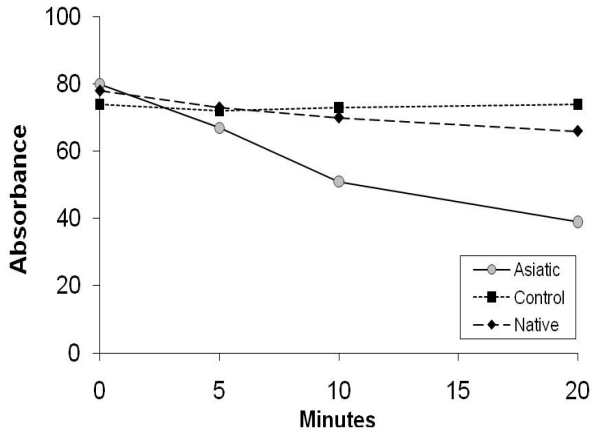

Figure 7. Bacterial clearance in invasive Asiatic clams and Native clams compared to control (no clam present).

Results for Micrographs Showing Cirri spacing

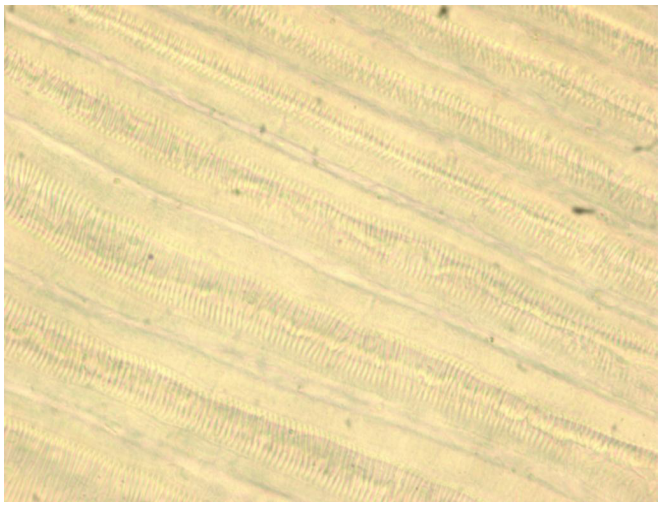

Figure 8. Microscopic image of cirri from *Corbicula* gills. Student measurements typically find cirri spacing of ~1 mm in *Corbicula*, and ~2 mm in native clams.

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Citing This Article

Graber Neufeld, D.S., and J.M. Yoder. 2011. The Role of Feeding Adaptations in Resource Competition between Invasive and Native Clams. Pages 78-87, in *Tested Studies for Laboratory Teaching,* Volume 32 (K. McMahon, Editor). Proceedings of the 32nd Conference of the Association for Biology Laboratory Education (ABLE), 445 pages.
http://www.ableweb.org/volumes/vol-32/?art=7